Interferon-γ-Mediated Allograft Rejection Exacerbates Cardiovascular Disease of Hyperlipidemic Murine Transplant Recipients

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Rationale: Transplantation, the most effective therapy for end-stage organ failure, is markedly limited by early-onset cardiovascular disease (CVD) and premature death of the host. The mechanistic basis of this increased CVD is not fully explained by known risk factors.

Objective: To investigate the role of alloimmune responses in promoting CVD of organ transplant recipients.

Methods and Results: We established an animal model of graft-exacerbated host CVD by combining murine models of atherosclerosis (apolipoprotein E–deficient recipients on standard diet) and of intra-abdominal graft rejection (heterotopic cardiac transplantation without immunosuppression). CVD was absent in normolipidemic hosts receiving allogeneic grafts and varied in severity among hyperlipidemic grafted hosts according to recipient–donor genetic disparities, most strikingly across an isolated major histocompatibility complex class II antigen barrier. Host disease manifested as increased atherosclerosis of the aorta that also involved the native coronary arteries and new findings of decreased cardiac contractility, ventricular dilatation, and diminished aortic compliance. Exacerbated CVD was accompanied by greater levels of circulating cytokines, especially interferon-γ and other Th1-type cytokines, and showed both systemic and intralesional activation of leukocytes, particularly T-helper cells. Serological neutralization of interferon-γ after allotransplantation prevented graft-related atherosclerosis, cardiomyopathy, and aortic stiffening in the host.

Conclusions: Our study reveals that sustained activation of the immune system because of chronic allogenrecognition exacerbates the atherogenic diathesis of hyperlipidemia and results in de novo cardiovascular dysfunction in organ transplant recipients. (Circ Res. 2015;117:943-955. DOI: 10.1161/CIRCRESAHA.115.306932.)

Key Words: atherosclerosis ■ cardiovascular disease ■ interferons ■ lymphocytes ■ transplantation
traditional risk factors, such as hypertension, hyperlipidemia, and diabetes mellitus, because of their common occurrence in patients with chronic kidney diseases before transplantation and the untoward effects of immunosuppressive drugs after transplantation. Although partially true, the Framingham Risk Score, based on these traditional risk factors and validated to predict future cardiovascular events in the general population, consistently and substantially underpredicts CVD morbidity and mortality in the kidney transplant population. The restricted predictive value of traditional risk factors for CVD in transplant recipients is interpreted to indicate the effect of additional, unidentified disease precipitants. Instead of Framingham criteria, the strongest risk factors of CVD events in renal transplant patients are pre-existing atherosclerosis and indices of poor graft function.

In turn, the most common cause of graft dysfunction is chronic rejection. Further evidence for immunologic causation is that circulating markers of inflammation, such as interleukin (IL)-6, C-reactive protein, and erythropoietin, are independently associated with CVD in renal transplant recipients. Premature CVD in renal transplant recipients has diverse clinical presentations. Although myocardial infarctions are common complications, an even greater increase in incidence has been noted in heart failure and sudden (presumed arrhythmic) cardiac deaths. This skewed presentation of CVD is compared with those in controls. Surgical procedures were performed with 6-to 12-week-old donor mice and 30-week-old recipient mice. Certain recipients were treated with neutralizing monoclonal antibody to IFN-γ (clone R4-6A2, BioXCell, West Lebanon, NH) or an isotype-matched, irrelevant rat IgG1 antibody (clone HRPN, BioXCell) at a loading dose of 250 µg SC 1 day before followed by 125 µg SC 3x per week, from 0 to 12 weeks after transplantation. The animals were euthanized at 42 weeks of age for analysis.

Surgical Procedures
Intra-abdominal heterotopic cardiac transplantation was performed by end-to-side anastomosis of the donor ascending aorta and pulmonary trunk to the recipient infrarenal aorta and inferior vena cava, respectively. The animals were observed daily after surgery; severely ill recipients (all of whom had rejected their grafts, except for 1 minor-mismatched recipient) were euthanized and adjudicated as deaths. Graft rejection was determined by abdominal palpation of the donor heartbeat. Alternatively, full thickness donor skin grafts were engrafted onto the back of recipient mice and covered with gauze and a securing bandage for 7 days. The animals were observed daily after surgery. Graft rejection was defined as necrosis >80% surface area. All animal studies were approved by Yale University’s Institutional Animal Care and Use Committee.

Analytic Techniques
The experimental methods are described in the Online Data Supplement.

Statistical Analysis
Data represent mean±SEM. Unpaired Student t test was used for comparisons between 2 groups, 1-way ANOVA was used for comparisons between >2 groups, and the Log-rank test was used to compare survival curves. Differences with 2-tailed P values of <0.05 were considered to indicate statistical significance. Statistical analyses were performed using Prism 4.0 (GraphPad Software, La Jolla, CA).

Results
Experimental Model of Graft-Related CVD
To study the pathogenesis of CVD in transplant recipients, we combined experimental models of atherosclerosis and graft rejection. We used apolipoprotein E-deficient (ApoE−/−) mice at 30 weeks of age as hosts to simulate the clinical risk factor of pre-existent atherosclerosis, regular chow to avoid anti-Th1 effects of high-fat diets, and minor-mismatched grafts as the transplanted organ for technical convenience and ease of monitoring function, no immunosuppression to permit allograft rejection and exclude confounding pharmacological effects, and 12 weeks of surveillance after transplantation to enable alloimmune responses to modulate host CVD (Figure 1A). Although major histocompatibility complex (MHC) and minor histocompatibility antigens expressed by the allograft may elicit immune rejection, it is not known which, if any, of these responses may elicit host CVD. Therefore, we screened several donor–recipient genetic disparities; controls included untransplanted and syngeneic-grafted animals. All the operations were technical successes (defined as host survival >3 days with beating cardiac grafts);
however, several of the recipients died unexpectedly or were euthanized because of poor health after their allografts had rejected between 6 and 12 weeks post operation (Figure 1B). In contrast, there were no deaths of untransplanted or syngeneic-grafted ApoE−/− mice. The deaths of ApoE−/− recipients with allogeneic grafts were unexpected as normolipidemic recipients with similar antigen-mismatched grafts are known to survive long term.27 Autopsies did not reveal direct graft causes of death, for example, hemorrhage or thrombosis. As expected, ApoE−/− male recipients acutely rejected full haplotype-mismatched hearts, chronically rejected either single MHC class II or class I antigen–mismatched hearts, and did not reject syngeneic hearts (Figure 1C and 1D). In addition, ApoE−/− female recipients of H-Y minor histocompatibility antigen-mismatched male hearts exhibited less graft attrition with fewer histological signs of rejection than for isolated MHC antigen differences (Online Figure I). Henceforth, the recipient groups are abbreviated as full-, class II-, class I-, or minor-mismatched recipients. Cholesterol and triglyceride serum levels did not differ between the various donor–recipient combinations (Figure 1E and 1F). Thus, the transplanted hosts with similar degrees of hyperlipidemia were suitable to examine for graft-related differences in CVD.

Graft Rejection Is Associated With Increased Atherosclerosis of the Host

Animals were euthanized at 12 weeks post operation (or earlier if distressed) to assess the extent of host atherosclerotic

![Figure 1. Experimental model of graft-related cardiovascular disease (CVD). A, ApoE−/− hosts fed regular chow received a heterotopic graft at 30 weeks of age without immunosuppression, and recipient cardiovascular function and disease were assessed at 12 weeks post operation (post-op). B, Survival of C57BL/6 ApoE−/− male mice after no transplantation (Untxp) or after transplantation with syngeneic (Syn; C57BL/6, H2b), full-mismatched (BALB/c, H2d), class II-mismatched (bm12, H2-Abm12), and class I-mismatched (bm1, H2-Kbm1) heterotopic cardiac grafts; P<0.05. C, Gift survival as determined by daily abdominal palpation for cardiac pulsation; P<0.001. D, Representative photomicrographs of the donor grafts at 12 weeks post operation showing cardiomyocyte destruction with cellular infiltrates (arrows, hematoxylin and eosin stain) and coronary artery neointima formation (insets, arrowheads, elastic Van Gieson stain); bars, 100 μm. Total cholesterol (E) and triglyceride (F) serum levels of hosts at 8 to 12 weeks post operation; n=4; P=0.23 and P=0.29, respectively.

disease. Aortic lipid deposition as measured by oil red O staining was 2- to 3-fold greater after allogeneic than syngeneic grafting or no transplantation (Figure 2A and 2B). Strikingly, the atherosclerotic lesions extended into the proximal segments of native coronary arteries, most consistently in recipients of class II–mismatched grafts (Figure 2C and 2D). In an additional group of animals in which the entire aorta was not processed for en face lipid staining, larger neointimal lesions were also found in cross sections of the aortic root in recipients of class II–mismatched versus syngeneic grafts (Figure 2E and 2F). The atherosclerotic plaques of allogeneic recipients were characterized by greater mineralization on von Kossa staining, a feature of advanced disease (Figure 2G and 2H). Increased calcification was confirmed by Alizarin red staining (not shown). Recipients of minor-mismatched grafts exhibited a lesser 1.5- to 2-fold increase in aortic atherosclerosis and no coronary artery lesions (Online Figure I). In contrast, allograft rejection in the absence of hyperlipidemia was not sufficient to induce atherosclerotic plaques in the aorta (nonspecific staining at the anastomotic site notwithstanding) or coronary

Figure 2. Graft rejection exacerbates pre-existing atherosclerosis of the host. ApoE−/− mice on standard diet were either not grafted or received heterotopic cardiac grafts of varying genetic disparities at 30 weeks of age, and atherosclerotic disease of the recipient was analyzed after 8 to 12 weeks. Representative oil red O stains of host aortas (A) and expressed as % of total area (B). Representative elastic Van Gieson (EVG) stains of host coronary arteries showing neointima (arrowhead; C) and expressed as intima to media area index (D). Representative EVG stains of host aortic roots showing neointima (arrowheads; E) and measured as intima area (F). Representative von Kossa stains of host aortic roots and brachiocephalic arteries (insets) showing mineralization (arrowheads; G) and expressed as % of plaque area (H). Bars, 200 μm. Data were pooled from 3 independent experiments; Untxp, Full, and Class I: n=3 to 4; Syn and Class II: n=7 to 12; *P<0.05, **P<0.01, and ***P<0.001 vs Untxp or Syn (there were no significant differences between the Untxp and Syn control groups or between the Full, Class II, and Class I allogeneic groups).
arteries of wild-type hosts (Online Figure II). Furthermore, class II–mismatched skin grafts, which underwent more rapid tissue necrosis within 2 weeks, only minimally increased aortic lipid deposition without coronary artery lesions in ApoE−/− recipients (Online Figure III). These data suggest that both host (pre-existent vascular disease) and graft (persistent alloimmune responses) factors are required to produce the accelerated atherosclerosis phenotype of our model.

Graft Rejection Induces De Novo Cardiovascular Dysfunction of the Host

Before euthanasia, cardiac and aortic functions of anesthetized hosts were assessed by echocardiography (Figure 3A). Indices of left ventricular systolic function, such as fractional shortening, were significantly decreased in recipients of class II–mismatched versus syngeneic grafts, although stroke volume was preserved (Figure 3B and 3C). Cardiac output was maintained (not shown) at the expense of ventricular dilatation, particularly at end systole (Figure 3D). Aortic size was also increased in recipients of allogeneic versus syngeneic grafts, particularly at end diastole (Figure 3E). These diameter changes translated as diminished aortic distension and recoil during the cardiac cycle (Figure 3F). Alterations in cardiovascular function were confirmed by invasive monitoring with a high-fidelity pressure sensor in a subgroup of animals. Compared with hosts of syngeneic grafts, recipients of class II disparate grafts had increased systolic blood pressure (Figure 3G) but not diastolic blood pressure (69.9±5.6 versus 48.5±6.6 mm Hg; P=0.07), pulse pressure (30.3±2.6 versus 27.2±2.0 mm Hg; P=0.38), or heart rate (327±10 versus 348±39 min−1; P=0.64). Peak rates of left ventricular pressure change were similar under basal conditions, but the allogeneic hosts demonstrated blunted inotropic and lusitropic responses to dobutamine infusion at 4 µg kg−1 min−1 (17.7±8.5% versus 55.8±6.7% increase in dP/dt max; P<0.05 and 6.4±10.7% versus 66.5±12.9% increase in dP/dt min; P<0.05). Calculation of arterial compliance based on distension and blood pressure measurements confirmed increased aortic stiffness in ApoE−/− recipients of class II–mismatched grafts (Figure 3H). Cardiovascular dysfunction was not apparent in ApoE−/− recipients of minor-mismatched cardiac grafts or class II–mismatched skin grafts and in normolipidemic wild-type recipients of class II–mismatched cardiac grafts, except for diminished responses to dobutamine infusion in the latter.

Figure 3. Graft rejection induces de novo cardiovascular dysfunction of the host. A, Representative M mode echocardiography of the native heart in ApoE−/− recipients of syngeneic (Syn) and class II–mismatched grafts at 12 weeks of post operation. Echocardiographically determined (B) fractional shortening, (C) stroke volume, (D) left ventricular (LV) internal dimensions, (E) ascending aorta diameters, and (F) ascending aorta distension of the host after syngeneic, full-, class II-, and class I–mismatched grafts; data were pooled from 3 independent experiments; Full and Class I; n=2; Syn and Class II; n=8 to 11; *P<0.05, **P<0.01, and ***P<0.001 vs Syn. Invasively determined central systolic blood pressure (G) and aortic compliance (ie, distension/ pulse pressure; H) in recipients of syngeneic and class II–mismatched grafts; data from a single experiment: n=3; *P<0.05 and **P<0.01 vs Syn.
group (Online Figures I and III). Thus, cardiac and thoracic aorta dysfunction of the hyperlipidemic host correlated with chronic allograft rejection in a remote body compartment.

Graft-Exacerbated CVD Is Associated With Systemic and Cardiovascular Inflammation

To determine how donor graft rejection within the abdomen may modulate host CVD in the thorax, we examined for evidence of circulating proinflammatory cytokines using multiplex bead-based immunoassays. Elevated plasma levels of the prototypic Th1 cytokine, IFN-γ, and the prototypic Th17 cytokine, IL-17, were found in recipients of class II–mismatched grafts compared with syngeneic-grafted or untransplanted ApoE−/− hosts at 12 weeks post operation (Figure 4A). This was associated with increased plasma levels of the IFN-γ inducers, IL-2 and IL-12, as well as the IL-17 inducers, IL-1β and IL-6 (Figure 4A). IL-12 p40 (a component of both IL-12 and the IL-17 inducer, IL-23) was also upregulated, but there were undetectable levels or no significant changes in multiple other cytokines assayed, including Th2, Th3, and Th9 lymphokines and macrophage growth factors (Online Figure IV). IFN-γ was also significantly elevated in full- and class I–mismatched recipients and trended to significant elevation in minor-mismatched recipients, but other changes of Th1- and Th17-related cytokines did not show consistent elevation across all allograft combinations (Figure 4A; Online Figure I). Because class II–mismatched recipients showed the greatest induction in circulating cytokines, we further examined the cardiovascular system of these hosts for inflammatory sequelae. A biological response to circulating cytokines could be inferred from increased expression of IFN-γ-inducible MHC class II antigens by host coronary artery endothelial cells.

Figure 4. Systemic and cardiovascular inflammation in recipients of rejecting allografts. A, Plasma levels of interferon (IFN)-γ, interleukin (IL)-2, IL-12 p70, IL-17, IL-1β, and IL-6 in ApoE−/− recipients of syngeneic (Syn) or full-, class II-, and class I–mismatched grafts at 12 weeks post operation; n=3 to 8; *P<0.05, **P<0.01, and ***P<0.001 vs Untxp and Syn. B, Representative images of immunofluorescence analysis of native coronary arteries in recipients of syngeneic (top) and class II–mismatched (bottom) grafts for the endothelial marker, CD31 (red color), for IFN-γ-inducible I-A/I-E MHC class II antigens (green color), and overlayed together with DAPI nuclear staining (blue color); bars, 50 µm. Host coronary arteries in allografted animals have partially occluded lumens and increased endothelial expression of I-A/I-E antigens.
Although circulating levels of immunoglobulin were unchanged, antibody deposition was noted in the host myocardial vasculature, and we documented that the expression of autoantigens by cultured vascular cells was upregulated by IFN-γ (Online Figure V). Finally, greater infiltrates of mononuclear leukocytes, particularly of T cells, were noted in host atherosclerotic aortic lesions in situ by immunofluorescence analysis, whereas sparse infiltrates of granulocytes were similar to controls (Online Figure VI).

**Graft-Exacerbated CVD Is Associated With Systemic and Intralesional T-Cell Activation**

We further characterized the phenotype of leukocytes isolated from ApoE−/− mice that received class II–mismatched versus syngeneic grafts by flow cytometry. At 12 weeks after allotransplantation, the frequency of circulating lymphocytes, in particular CD4+ T-helper cells, was increased (Figure 5A). Furthermore, these cells displayed a greater expression of the activation markers, CD25 and CD69 (Figure 5B). Interestingly, the CD25high/CD4+ subpopulation, typically regulatory T cells, did not increase in frequency or acquire CD69 expression. The activation of circulating CD4+ T cells was far less apparent at 4 and 8 weeks post operation (Online Figure VII). The number of circulating activated CD4+ T cells was only modestly increased in normolipidemic hosts of cardiac allografts and unchanged in hyperlipidemic hosts of skin allografts (Online Figures II and III). Similar to the findings in the circulation, the proportion of activated CD4+ T cells was higher in the spleens of ApoE−/− recipients bearing class II–mismatched grafts (Figure 5C and 5D). Notably, there were also more CD4+ T cells with greater expression of CD25 and CD69 infiltrating the native aorta of these animals (Figure 5E and 5F). The differences in activated T-helper cells were even more significant considering that 7-fold more CD45+ leukocytes were isolated from the aortas of allogeneic than syngeneic recipients (Figure 5G). There was also an increased frequency and total number of activated (CD25+/CD69+) CD8+ cytotoxic T cells, inflammatory (CCR2+/Ly6Chigh) CD11b+ monocytes, and proatherogenic (CD5−/CD43−) B220+ B-2 cells (Online Figures VIII and IX); however, these were of lesser magnitude than the changes in CD4+ T cells. On the other hand, the frequency of patrolling (CCR2−/Ly6Clo) monocytes, atheroprotective (CD5+/CD43+) B-1a cells, and Ly6G+ neutrophils were lower in allografted animals (Online Figures VIII and IX). Together, the elevated levels of circulating cytokines and increased number of activated leukocytes demonstrated greater activation of the immune system in hyperlipidemic recipients of rejecting allografts that correlated with the occurrence of exacerbated host CVD.
IFN-γ Promotes Graft-Exacerbated CVD in the Transplant Recipient

Finally, we asked whether IFN-γ could account for the increased severity of CVD in ApoE−/− mice receiving an allograft as, in addition to higher plasma levels, synthesis of this key proarteriosclerotic factor by CD4+ T cells in the circulation and secondary lymphoid tissues was disproportionately increased (30- to 50-fold more Th1 cells compared with 6-fold more Th17 cells) in recipients of class II–mismatched versus syngeneic grafts (Figure 6A and 6B). There was also increased production of IFN-γ by leukocytes other than CD4+ T cells (but of lesser magnitude than in Th1 cells) and in production of the IFN-γ-inducers, IL-12 and IL-18, within the host heart (Online Figure X). Administration of neutralizing antibody to IFN-γ after allotransplantation diminished systemic Th1 (but increased Th17) differentiation, reduced the levels of circulating IFN-γ, and decreased the expression of IFN-γ inducers, IFN-γ, and IFN-γ-inducible molecules in the host heart.

Figure 6. Effects of interferon (IFN)-γ neutralization on immune and inflammatory responses. ApoE−/− recipients of syngeneic (Syn) and class II–mismatched grafts were treated with irrelevant IgG or IFN-γ antibody from 0 to 12 weeks post operation. A, Flow cytometry of IFN-γ and interleukin (IL)-17 expression by circulating CD4+ T cells. B, Similar analyses in splenic CD4+ T cells. Cells for each analysis were pooled from 3 animals. ELISA determination of IFN-γ plasma levels (C) and quantitative reverse transcription polymerase chain reaction analysis of recipient hearts for IFN-γ and IFN-γ-inducible CXCL10 transcripts (D); n=3 to 5 per group; *P<0.05 and **P<0.001 vs other groups. E, Immunofluorescence analysis of native coronary arteries for the endothelial marker, CD31 (red color), for IFN-γ–inducible I-A/I-E MHC class II antigens (green color), and overlayed together with DAPI staining of nuclei (blue color); bars, 50 µm. IFN-γ neutralization (bottom) decreased inducible I-A/I-E antigen expression by arterial endothelial cells but not the constitutive expression by perivascular leukocytes.
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(Figure 6A and 6E), as well as the donor graft (Online Figure X). As expected, neutralization of IFN-γ reduced the extent of arteriosclerosis in the vessels of the graft (Online Figure XI). Strikingly, anti–IFN-γ therapy also prevented the allograft-dependent increase in lipid deposition into and intimal thickening of the host aorta (Figure 7A–7D) without causing a reduction in circulating lipids (Online Figure XI). Moreover, this treatment strategy prevented left ventricular dilatation, preserved left ventricular contractility, and restored aortic distension in allograft recipients (Figure 7E–7H). Despite these benefits, neutralizing IFN-γ did not ensure survival of all hosts receiving allografts (Online Figure XI), although the experiment was not adequately powered to test for a partial improvement in survival.

Discussion

We find that pre-existent vascular disease and graft rejection are associated with accelerated atherosclerosis and de novo cardiovascular dysfunction in murine transplant recipients. Furthermore, IFN-γ produced by alloimmune responses and consequent inflammation plays a nonredundant role in promoting the graft-exacerbated atherosclerosis, cardiomyopathy,

![Image](https://example.com/image.png)

**Figure 7.** Interferon (IFN)-γ neutralization prevents graft-exacerbated host cardiovascular disease. ApoE−/− recipients of syngeneic (Syn) or class II–mismatched grafts were treated with irrelevant IgG vs neutralizing antibody to IFN-γ for 12 weeks post operation. Representative oil red O stains of host aortas (A) and expressed as % of total area (B). Representative elastic Van Gieson stains of host aortic roots showing neointima (arrowheads), bar=200 μm (C) and intima area (D). Left ventricular (LV) internal dimensions (E), LV fractional shortening (F), ascending aorta diameters (G), and ascending aorta distension (H) during the cardiac cycle derived from echocardiographic images of the host heart and aorta. n=3 to 6 per group; *P<0.05 and **P<0.01 vs other groups.
and aortic stiffening of ApoE−/− hosts. The manifestations of CVD associated with graft rejection are qualitatively different from those of the enhanced atherosclerosis and modulation of plaque composition that has been described in hyperlipidemic mice with several other systemic inflammatory stimuli of autoimmune disease, microbial infection, and tissue injury.20–31 In addition to greater lipid deposition, larger neointima formation, more extensive distribution, and increased mineralization of atherosclerotic lesions, we also find decreased ventricular contractility, ventricular dilatation, diminished contractile reserve, systolic hypertension, aortic stiffening, and unexplained deaths. This constellation of atherosclerosis and cardiovascular abnormalities resembles the skewed clinical presentation of graft-related CVD with accelerated heart failure and sudden death in renal transplant patients.8,20–22

By comparing hyperlipidemic with normolipidemic hosts, we conclude that pre-existent atherosclerosis because of conventional disease precipitants is necessary for the exaggerated inflammation and disease manifestations of the transplantation-induced phenotype. By varying donor–recipient genetic disparities and comparing different types of grafts, we conclude that both the strength and chronicity of alloimmune responses affect the pathogenesis of host CVD. Full-mismatched hearts are vigorously rejected within a relatively short period of time, and alloimmune responses, deprived of antigen, begin to resolve after the graft is destroyed. Similarly, class II–mismatched skin grafts rapidly necrose because of failure of secondary vascularization. On the other hand, less vigorous rejection of class I– and minor-mismatched hearts is expected to delay the initiation of systemic inflammation in the host. The greatest degree of graft-exacerbated CVD (as indicated by significant coronary atherosclerosis and cardiac dysfunction) in recipients of class II–mismatched hearts may reflect the combination of intermediate strength, but relatively persistent alloresponses known to result in robust chronic graft rejection.27 Alternatively, the prominent sequelae of class II–mismatched grafts may point to a pivotal role for CD4+ T cells and their cytokines in the pathogenesis of graft-exacerbated CVD.

We postulate 3 mechanisms of disease in our model of graft-rejection-induced inflammation and disease manifestations of the transplantation-induced phenotype. First, systemic inflammation may enhance proatherosclerotic factors, we do not discount immunologic perturbation of traditional metabolic and hemodynamic risk factors contributing to the pathogenesis of graft-exacerbated CVD in our model. Moderate hypertension (39% increase in mean blood pressure under conditions of anesthesia) ensued in recipients of class II–mismatched grafts at 12 weeks of postoperation. However, this degree of blood pressure elevation is not thought to result in phenotypic changes in ApoE−/− mice.32 In addition, minor changes in circulating lipids that did not reach statistical significance are unlikely to account for the qualitative differences in CVD manifestations after allotransplantation.

We find evidence for activation of diverse immune responses in ApoE−/− transplant recipients, including cell-mediated and humoral adaptive immunity, as is well described for the development of atherosclerosis in hyperlipidemic mice and for allograft rejection by normolipidemic hosts across the same strain combinations.33–36 We extend these previous findings by demonstrating a marked enhancement of systemic inflammation, particularly of circulating CD4+ T-cell activation, when the 2 models of disease are combined. Innate immune cells also play an important role in atherosclerosis and graft rejection.37,38 The increased total number of macrophages with a skewed inflammatory phenotype within atherosclerotic lesions of ApoE−/− hosts may reflect activation by adaptive immune responses or by recognition of tissue damage signals in rejecting allografts similar to the injury effect seen with myocardial infarction of native hearts.31 In this initial study, we focused on the role of T-helper cells and IFN-γ that are crucial activators of many components of the immune system. Thus, other immune effectors dependent on CD4+ T cells and IFN-γ are also likely to contribute to graft-exacerbated CVD and will be the subject of future studies. The actions of T-helper cells and IFN-γ in the pathogenesis of atherosclerosis have been extensively studied, as discussed below. However, we have not yet determined the exact mechanism(s) for cardiac failure and aortic stiffening. Although IFN-γ is necessary for these pathological changes in our model, it may not be sufficient. Further work is required to determine whether cardiovascular dysfunction is solely a consequence of atherosclerosis, that is, after a continuum of occlusive arterial disease, myocardial infarction, and heart failure.39 In other experimental systems, autoreactive T cells, autoreactive B cells, autoantibodies, and even individual cytokines can directly impair cardiac function independent of atherosclerotic disease.40–42 Acute administration of endotoxin, a bacterial product that triggers an intense inflammatory response, is known to depress left ventricular systolic function independent of preload or afterload and result in greater systolic than diastolic left ventricular dilatation.43 T cells and their cytokines may also result in stiffening of nonatherosclerotic aortas,44 and there is clinical evidence that aortic stiffness may induce cardiac dysfunction.45

We postulate 3 mechanisms of disease in our model of graft-exacerbated CVD, which are not mutually exclusive. First, systemic inflammation may enhance proatherosclerotic immune responses within the artery wall representing an endocrine effect of graft rejection on host disease. In support of this hypothesis, increased IL-6 levels have been found in patients with graft-related CVD,48 although measures of other Th1- and Th17-related cytokines have not been reported in
this particular clinical scenario. We have previously shown an inflammatory pathway of IFN-γ production by T cells in response to IL-12 and IL-18, independent of their cognate receptor interactions, in human coronary atherosclerosis and that IFN-γ is necessary and sufficient for the progression of human coronary arteriosclerosis. IL-17, another key effector cytokine of adaptive immune responses, is produced concomitantly with IFN-γ by human coronary artery–infiltrating T cells with synergistic effects on vessel wall inflammation but does not contribute to neointima formation. Proatherosclerotic effects of IFN-γ and, controversially, of IL-17 have also been described in murine models and further work is required to dissect their interactions in graft-exacerbated CVD. Second, alloimmune responses between host and graft may directly damage recipient arteries via paracrine mechanisms. Host antitumor T cells may recirculate from the transplanted organ to the native heart and vasculature and contribute to pathological inflammatory responses, in particular if donor passenger leukocytes have taken resident in recipient tissues. Graft antihost T cells are known to target the endothelium of transplant recipient microvessels causing clinical disease, and similar unrecognized responses may occur in conduit arteries as these vessels are not generally sampled in diagnostic procedures. Third, graft rejection may induce autoimmune injury as the alloimmune response spreads to encompass antigens shared by the graft and the host. Of relevance, autoantibodies to nuclear and cytoplasmic self-antigens have been characterized in C57BL/6 recipients of bm12 cardiac grafts that result in donor artery injury, although effects on the normolipidemic host vasculature have not been described. In the latter study, autoantibody production by recipient B cells was dependent on donor CD4+T cell help, and similar interactions may be operational for graft-exacerbated CVD of the host.

A limitation of our experimental system is the absence of immunosuppressive agents, unlike the clinical scenario of graft-related CVD. Effective immunosuppression in rodents readily leads to allograft acceptance with little residual immune responses. This may be because rodent models are primarily dependent on naive T-cell activation, which is more easily suppressed than memory responses that dominate the early response to human allografts. Alternative approaches to obtain attenuated graft rejection by subtherapeutic doses or transient administration of drugs would invoke off-target effects on the host. Many immunosuppressive agents have unfavorable side effects on CVD risk factors and may promote atherosclerosis. Conversely, immunosuppressants may inhibit the inflammation-driven progression of atherosclerosis.

We reasoned that avoiding confounding immunosuppression and depending on controlled immunogenetic differences were preferable for our initial studies. It may be that our experimental system best models the clinical situation in which immunosuppressants are discontinued after kidney graft failure, and dialysis is reintroduced without excision of the donor organ (that is often contraindicated because of the risks of reoperative surgery in the inflamed site). With the reintroduction of immune competency, unrestrained alloimmune responses are expected and, significantly, are associated with a 3-fold spike in CVD morbidity and mortality of renal transplant recipients.

Even after correcting for the selection bias of dialysis patients listed for transplantation, a deleterious effect for a failed allograft was revealed compared with no previous transplant. Clinicians have focused attention on the chronic inflammatory state resulting from failed kidney allografts, and some strongly recommend graft nephrectomy or continued use of immunosuppressive drugs in this setting, although there is no consensus for the management of this difficult problem.

Medical advances have greatly improved the early but not late outcomes after all types of organ transplantation. Chronic rejection and CVD have emerged as the leading obstacles to graft and patient survival. Both processes are characterized by arteriosclerotic lesions and ischemic and fibrotic sequelae, both diseases represent risk factors for each other. In our opinion, their concurrent manifestations in transplant recipients are not a coincidence but represent a failure of current immunosuppressive regimens to prevent and treat these conditions, unlike the favorable results for acute rejection. The immunologic mechanisms that contribute to the pathogenesis of native atherosclerosis and graft arteriosclerosis have been extensively studied. As discussed above, we and others have shown that adaptive immune responses, in particular that of Th1 cells, play a central role in both processes. Thus, common pathogenetic mechanisms of excessive IFN-γ activity on vessel wall cells may link graft arteriosclerosis and host CVD. Although our study adds to the rationale for neutralizing IFN-γ as part of the management of transplant recipients, the explained failure of this therapy to prevent all host deaths in our model despite improved cardiovascular outcomes indicates that the adverse effects of having chronic immune stimulation caused by the presence of allogeneic tissue is likely to be more complex than the effects of increased levels of a single cytokine.

In conclusion, our experimental model replicates many aspects of a disease process that represents a major obstacle to the long-term success of organ replacement therapy. It provides a preclinical framework to define hitherto unsuspected disease mechanisms and to test new therapeutic approaches to prevent or reverse graft-related CVD of the transplant recipient. Furthermore, our work identifies IFN-γ as a therapeutic target to inhibit CVD of the host in addition to previous studies that have validated IFN-γ antagonism as treatment for atherosclerosis of the graft.

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Disclosures
None.

References


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Novelty and Significance

What Is Known?
- Organ transplant recipients have an increased risk of cardiovascular disease (CVD) and premature death.
- The increased occurrence of CVD and its complications is only partially accounted by traditional risk factors.
- The restricted predictive value of traditional risk factors for CVD in organ transplant recipients is interpreted to indicate the effect of additional, unidentified disease precipitants.

What New Information Does This Article Contribute?
- Graft rejection in hyperlipidemic mice exacerbates atherosclerosis and induces a neo-dysfunction of the recipient heart.
- Increased atherosclerosis and cardiovascular dysfunction of the host are associated with systemic and local activation of the immune system, particularly of T-helper cells and the production of interferon (IFN)-γ.
- Neutralization of IFN-γ prevents the worsening of atherosclerosis and the onset of cardiovascular dysfunction in hosts with rejecting grafts.

Solid organ transplantation is life-saving but may cause disease and death of the recipient if it fails, is rejected, or, in rare circumstances, transmits malignant cells, pathogens, or passenger leukocytes sufficient for graft-versus-host disease. A detrimental effect of the graft on the host cardiovascular system has not been suspected although graft dysfunction is an independent risk factor of CVD in organ transplant recipients. In hyperlipidemic mice, we find that intra-abdominal graft rejection is associated with increased atherosclerosis and dysfunction of the host heart and aorta. This disease association across body cavities is characterized by systemic and local activation of leukocytes and the production of cytokines, particular of T-helper cells and IFN-γ. Antibody neutralization of IFN-γ prevents the exacerbated host atherosclerosis and cardiovascular dysfunction. Our findings suggest that graft-related inflammation in organ transplant recipients may worsen their CVD and lead to their premature death. In addition to the known benefits of inhibiting IFN-γ responses in native atherosclerotic disease and in chronic allograft vasculopathy, we identify IFN-γ as a therapeutic target in graft-exacerbated CVD of the host. Our data provide a rationale for extending ongoing treatment trials with anti–IFN-γ antibody from patients with autoimmunity to allograft recipients. 
Interferon-γ–Mediated Allograft Rejection Exacerbates Cardiovascular Disease of Hyperlipidemic Murine Transplant Recipients

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Supplemental Materials

Detailed Methods

Histology

Anesthetized animals were exsanguinated for serology, euthanized by perfusion through the left ventricle with saline until atrial effluents were clear, and the specimens were excised. The base of the host heart with aortic root, the host brachiocephalic artery, and the donor heart were post-fixed in 4% paraformaldehyde for 24 h at 4 °C, and embedded in paraffin. Alternatively, specimens were immediately frozen in OCT. Hematoxylin and eosin (H&E), elastin Van Gieson (EVG), von Kossa, and Alizarin red stains were performed on 5 µm-thick sections by a research core facility of Yale’s Department of Pathology using standard techniques. Morphological analyses were performed in 3-5 sections by tracing the luminal, internal, and external elastic lamina perimeters to calculate intima and media areas using ImageJ software (National Institutes of Health, Bethesda, MD). For lipid detection, the aorta was dissected free of connective tissue and fat, excised, opened longitudinally, post-fixed in 4% paraformaldehyde, pinned onto a silicone plate luminal surface up, incubated with 0.2% Oil Red O solution (Sigma-Aldrich, St. Louis, MO) for 50 min at room temperature, destained in 78% methanol for 5 min, and then mounted on glass slides using aqueous mounting medium. Images were acquired using a Nikon SMZ 1000 microscope connected to a Kodak DC290 camera and multiple high power views were combined for each aorta. Lipid lesions were individually traced and the sum of their area to that of the whole aorta was quantified using ImageJ.

Cardiovascular assessment

Ventricular and aortic function were evaluated using the Vevo 770 echocardiography system (Visualsonic, Toronto, Canada). In brief, mice were anesthetized with isofluorane and cardiac and aortic images were acquired using 15-45 MHz (RMV707B) and 20-60 MHz (RMV704) transducers, respectively. Transthoracic M-mode images of the heart were obtained in parasternal short axis at the level of the papillary muscle to determine left ventricular dimensions, fractional shortening, and cardiac output. Additionally, B-mode images of the thoracic aorta were obtained in a longitudinal plane to measure end-systolic and end-diastolic diameters of the distal ascending aorta. In a subgroup of animals, cardiovascular function was also invasively evaluated using a high-fidelity 1.9 French transducer-tipped catheter (Millar Inc., Houston, TX). In brief, mice were anesthetized by ketamine injection, supported on a ventilator, and maintained on isofluorane inhalation. The transducer-tipped catheter was advanced into the left ventricle via the right carotid artery. Pressures and volumes were recorded under basal conditions and with gradually increasing doses of dobutamine infusion from 0-4 µg/kg/min. Data were recorded by MacLab software and then analyzed by the Heartbeat program.

Serological analyses

Blood was collected in Eppendorf tubes and serum was obtained after clot removal. Alternatively, blood from EDTA-coated tubes was separated into cellular and plasma components by centrifugation. Lipid levels were measured using Amplex Red Cholesterol Assay Kits (Invitrogen, Grand Island, NY) and Triglyceride Determination Kits (Sigma-Aldrich), respectively. IgG and IgM plasma levels were quantified using ELISA kits (Bethyl Laboratories, Montgomery, TX). Plasma cytokines were evaluated using a Luminex-based multicytokine kit (Millipore, Billerica, MA) according to the manufacturer’s protocol. In brief, specimens and standards were sequentially incubated with premixed antibody-coated bead sets, detection antibodies, and streptavidin-phycocerythrin. Reporter fluorescence of the beads was determined using a Luminex 200 analyzer, samples were run in duplicate and quantified based on a unique standard curve for each analyte. IFN-γ plasma levels were also determined in additional samples using a sandwich ELISA kit (Millipore) following the manufacturer’s instructions.
Flow cytometry

Blood was collected in EDTA-coated tubes, cells were separated by centrifugation, and red blood cells were lysed with ACK buffer. Splenocytes were obtained after mincing and straining tissue, centrifugation, and red blood cell lysis. For isolation of artery-infiltrating leukocytes, aortas were digested with 125 U/mL collagenase XI, 60 U/mL hyaluronidase I, 60 U/mL DNase 1, and 450 U/mL collagenase I (Sigma-Aldrich) in phosphate-buffered saline containing 20 mM HEPES at 37 °C for 1-2 h. The tissue and supernatant were resuspended in RPMI-1640 medium at 4 °C and passed through 0.5 and 0.1 mm sieves. Leukocytes were isolated with CD45 antibody-coated magnetic beads (Miltenyi Biotec, Auburn, CA) and cell separation columns; selected cells were >95% CD45+. Cell surface molecules were labeled with fluorescence-conjugated monoclonal antibodies to CD4, CD5, CD8, CD11b, CD25, CD43, CD45, CD69, B220, Ly6C, Ly6G (all from eBioscience, San Diego, CA), CCR2 (R&D, Minneapolis, MN), or isotype-matched, irrelevant IgG (eBioscience). For intracellular staining, the cells were treated with phorbol myristate acetate at 10 ng/mL and ionomycin at 1 µM for 5 h in the presence of brefeldin A at 10 µg/mL, labeled with antibodies to cell surface markers, fixed with 4% paraformaldehyde and permeabilized with 0.1% w/v saponin, and then labeled with PE-conjugated, rat anti-mouse IFN-γ, FITC-conjugated rat anti-mouse IL-17, or fluorescence-conjugated, isotype-matched, irrelevant antibodies (eBioscience). Additionally, cultured lung microvascular endothelial cells and aortic smooth muscle cells from C57BL/6 mice were incubated with serum (1:1,000 dilution) from various recipients and surface bound immunoglobulin was detected using rat anti-mouse IgG (Invitrogen). Analysis was performed using a LSR II system (BD Biosciences, San Jose CA) and FlowJo software (BD Biosciences).

Immunofluorescence microscopy

Five µm-thick sections of OCT-embedded heart specimens were fixed in acetone and sequentially labeled with rat anti-mouse CD31 monoclonal antibody (BD Biosciences) followed by Alexa Fluor 594-conjugated anti-rat IgG (Invitrogen), and then by Alexa Fluor 488-conjugated rat anti-mouse I-A/I-E monoclonal antibody (Biolegend, San Diego, CA). Alternatively, myocardium sections were labelled with antibodies to IgG (Invitrogen), CD31, and smooth muscle α-actin (Santa Cruz Biotechnology) and aortic root sections were labelled with antibodies to CD3, F4/80 (Abcam, Cambridge, MA), B220, and Ly6G (eBioscience). Controls included isotype-matched, irrelevant IgG. The specimens were mounted with ProLong Gold antifade reagent with DAPI (Invitrogen). Images were acquired using an Axiovert 200M microscopy system (Carl Zeiss MicroImaging, Thornwood, NY) with Volocity 6.3 software (PerkinElmer, Waltham, MA).

Real-time quantitative RT-PCR

Serial sections of OCT-embedded tissue were immersed in RLT lysis buffer (QIAGEN, Valencia, CA), vigorously vortexed, and total RNA was isolated using Rneasy mini kits and DNA digestion kits (QIAGEN) according to the manufacturer’s protocol. RT with random hexamer and oligo-dT primers was performed according to the Multiscribe RT system protocol (Applied Biosystems, Foster City, CA). RT-PCR reactions were prepared with TaqMan PCR Master Mix and predeveloped assay reagents from Applied Biosystems. Duplicate samples were analyzed on an iCycler (Bio-Rad Laboratories, Hercules, CA). RNA samples processed without the RT enzyme were used as negative controls. The expression of IFN-γ, CXCL10, IL12a, and IL-18 transcripts were normalized to GAPDH; a second reference gene of β-actin gave similar results.
Supplemental Figures and Figure Legends

Online Figure I. Single minor histocompatibility antigen mismatch. (A) Survival of 30 wk old C57BL/6 ApoE\(^{-/-}\) female mice either untransplanted (Untxp) or receiving syngeneic male heterotopic cardiac grafts, all grafts were beating at 12 wk post-op; \(n=4\), \(P=0.37\). (B) Representative photomicrographs of the native heart or of the donor graft in a minor-mismatched recipient at 12 wk post-op showing mild cellular infiltrates (arrowhead, H&E stain) and minimal neointima (inset, EVG stain), bars=100 µm. (C) Representative Oil Red O stains of the host aorta and expressed as % total area; \(n=3-4\), \(P=1.0\) vs. Untxp. (D) Representative EVG stains of host coronary artery frozen sections showing minimal neointima, bar=100 µm and expressed as intima to media area index; \(n=13-17\) coronary arteries of >50 µm diameter from 3-4 recipients, \(P=0.67\) vs. Untxp. (E) Left ventricular fractional shortening, left ventricular internal dimensions, and ascending aorta diameters at end-systole and end-diastole of female recipients of male grafts and male recipients of male grafts; \(n=3-4\), \(P>0.05\) vs. Syn. (F) Plasma levels of IFN-γ in untransplanted and minor-mismatched recipients; \(n=3-4\), \(P>0.05\) vs. Untxp.
Online Figure II. Normolipidemic recipients of class II-mismatched cardiac grafts. (A) Survival of 30 wk old C57BL/6 male mice after transplantation with syngeneic or bm12 male heterotopic cardiac grafts; n=8, P<0.001. (B) Representative photomicrographs of the donor grafts at 12 wk post-op showing cardiomyocyte destruction with cellular infiltrates (arrow, H&E stain) and coronary artery neointima formation (inset, arrowhead, EVG stain), bars=100 µm. (C) Representative Oil Red O stains of the host aorta and EVG stains of host coronary artery frozen sections at 12 wk post-op showing no lipid accumulation (besides nonspecific uptake at anastomotic sites, arrowheads) and no neointima, bar=100 µm. (D) Change in dP/dt\text{max} and dP/dt\text{min} from basal to stress conditions after dobutamine infusion at 4 µg/kg/min; n=4-8, **P<0.01 vs. Syn. (E) CD4 and CD8 expression by circulating mononuclear leukocytes at 12 wk post-op. (F) Circulating CD4\textsuperscript{+} T cells were further analyzed for CD25 and CD69 expression.
Online Figure III. MHC class II antigen disparate skin grafts to ApoE⁻/⁻ recipients. (A) Survival of 30 wk old C57BL/6 ApoE⁻/⁻ male mice after grafting with syngeneic or bm12 male skin; n=4, P<0.001. (B) Representative photographs of the grafts at 3 wk post-op showing rejection of allogeneic not syngeneic skin. (C) Representative Oil Red O stains of the host aorta and expressed as % of total area; n=4, *P<0.05 vs. Syn. (D) Left ventricular fractional shortening and left ventricular internal dimensions at end-systole and end-diastole at 12 wk post-op; n=4, P>0.05 vs. Syn. (E) CD4 and CD8 expression by circulating mononuclear leukocytes at 12 wk post-op. (F) Circulating CD4⁺ T cells were further analyzed for CD25 and CD69 expression.
Online Figure IV. Circulating levels of lymphocyte- and macrophage-derived cytokines. Plasma levels of IL-12 p40, IL-7, IL-15, IL-4, IL-5, IL-13, IL-9, IL-10, TNF-α, M-CSF, GM-CSF and G-CSF were measured by multiplex bead-based immunoassay in untransplanted ApoE^{-/-} mice or in ApoE^{-/-} recipients of syngeneic or full-, class II- and class I-mismatched grafts at 12 wk post-op; n=3-8, *P<0.05 vs. Untxp and Syn.
Online Figure V. Antibody deposition in host coronary arteries. (A) Plasma levels of IgM and IgG in ApoE−/− recipients of syngeneic and class II-mismatched grafts at 12 wk post-op; n=4, *P=0.062 and 0.75, respectively. (B) Representative images of immunofluorescence analysis of native coronary arteries from ApoE−/− recipients of syngeneic (upper panels) and class II-mismatched (lower panels) grafts for IgG (green color), CD31 (blue color), smooth muscle α-actin (red color), and overlay of images to demonstrate immunoglobulin deposition on host endothelium and neointimal (but not medial) smooth muscle cells in vivo, bars=50 µm. (C) Flow cytometric analysis to detect reactivity of IgG in serum (1:1,000 dilution) from wild-type C57BL/6 mice or ApoE−/− recipients of syngeneic and class II-mismatched grafts to autologous cultured endothelial cells and smooth muscle cells either untreated or treated with IFN-γ (10 ng/mL x 3 d) in vitro. Cytokine treatment increased the expression of cell surface molecules binding autoreactive serum antibodies present in ApoE−/− transplant recipients, particularly of class II-mismatched grafts.
Online Figure VI. Leukocytic infiltrates in atherosclerotic lesions of the host aorta. Representative images of immunofluorescence analysis of native aortic roots from ApoE<sup>-/-</sup> recipients of syngeneic (left panels) and class II-mismatched (right panels) grafts for (A) CD3 T cell marker (green color), (B) B220 B cell marker (red color), (C) F4/80 macrophage marker (red color), and (D) Ly6G granulocyte marker (green color) overlayed with DAPI nuclear staining (blue color) to differentiate aortic annular tissue (below) from lumen (above), bars=200 µm. The atherosclerotic lesions of allografted recipients demonstrated a greater infiltrate of mononuclear leukocytes, particularly of T cells, but a similar sparse granulocyte infiltrate to syngeneic grafted recipients.
Online Figure VII. T cell phenotype at different times after transplantation. Flow cytometric analysis was performed on circulating mononuclear leukocytes pooled from 3 ApoE−/− recipients of syngeneic or class II-mismatched grafts. CD4 and CD8 expression by CD45+ leukocytes at (A) 4 wk and (B) 8 wk post-op (corresponding dot-plots at 12 wk post-op are shown in Fig. 5). Further analysis of the CD4+ T cell population for CD25 and CD69 expression at (C) 4 wk and (D) 8 wk post-op (corresponding dot-plots at 12 wk post-op are also shown in Fig. 5). Temporal trend of (E) CD25 expression by circulating CD4+ T cells and (F) CD69 expression by circulating CD4+ T cells.
Online Figure VIII. Phenotype of cytotoxic T cells and monocyte/macrophages. Leukocyte phenotypes were characterized by flow cytometry in ApoE<sup>-/-</sup> recipients of syngeneic and class II-mismatched grafts at 12 wk post-op. Expression of the activation markers, CD25 and CD69 by CD8<sup>+</sup> T cells isolated from (A) blood, (B) spleen, and (C) host aorta. CD11b<sup>+</sup> monocyte/macrophages from (D) blood, (E) spleen, and (F) host aorta were also analyzed for the expression of the inflammatory markers, CCR2 and Ly6C to define subsets of CCR2<sup>-/Ly6C<sub>low</sub></sup> patrolling monocytes and CCR2<sup>+/Ly6C<sub>high</sub></sup> inflammatory monocytes.
Online Figure IX. Phenotype of B cells and neutrophils. Leukocyte phenotypes were characterized by flow cytometry in ApoE−/− recipients of syngeneic and class II-mismatched grafts at 12 wk post-op. Expression of the B cell marker, B220 (CD45R isoform) and the neutrophil marker, Ly6G by CD45+ leukocytes isolated from (A) blood, (B) spleen, and (C) host aorta. B220+ B cells from (D) blood, (E) spleen, and (F) host aorta were further analyzed for the expression of CD5 and CD43 to define subsets of CD5+/CD43+ B-1a cells, CD5-/CD43+ B-1b cells, and CD43-/CD5- B-2 cells.
Online Figure X. Effects of IFN-γ neutralization on immune and inflammatory responses.

(A) IFN-γ and IL-17 expression by circulating CD45⁺ leukocytes not expressing CD4 in ApoE⁻⁻ recipients of syngeneic and class II-mismatched grafts treated with irrelevant IgG or anti-IFN-γ antibody (cells pooled from 3 animals). (B) Similar analyses in splenic cells. (C) Quantitative RT-PCR of recipient hearts for IL-12 and IL-18 RNA. (D) Similar analysis of donor grafts for IFN-γ, CXCL10, IL-12, and IL-18 transcripts. n=3-5, *P<0.05, **P<0.01, ***P<0.001, vs. other groups.
Online Figure XI. Effects of IFN-γ neutralization on graft and host CVD. ApoE−/− recipients of syngeneic or class II-mismatched grafts were treated with irrelevant IgG or anti-IFN-γ antibody from 0-12 wk post-op. (A) Representative EVG stains of donor heart graft frozen sections at 12 wk post-op showing coronary artery neointima (arrowheads, bar=100 µm) and (B) expressed as a ratio of intima to media area; n=3-5, **P<0.01 vs. other groups. (C) Total cholesterol, (D) triglyceride, (E) HDL cholesterol, and (F) LDL cholesterol plasma levels of hosts at 8-12 wk post-op; n=3-5, P>0.05. (G) Host survival; n=3-6, P=0.18 for Class II IgG vs. Class II anti-IFN-γ and (H) graft survival; n=3-6, P=0.10 for Class II IgG vs. Class II anti-IFN-γ.